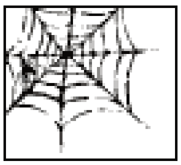
The Volunteer Monitor

The National Newsletter of Volunteer Water Quality Monitoring
Vol. 9, No. 1, Spring 1997



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Special Topic: Methods and Techniques

Rotating co-editors

The Volunteer Monitor has a permanent editor and volunteer editorial board. In addition, a different monitoring group serves as co-editor for each issue.

This issue was co-edited by Rhode Island Watershed Watch, a Cooperative Extension-sponsored program with over 200 volunteers monitoring lakes, ponds, and rivers in Rhode Island and Connecticut. The program, which is celebrating its 10th anniversary this year, can be reached at 210B Woodward Hall, Natural Resources Science Department, Kingston, RI 02881-0804; 401/874-2905.

The following articles appear in this edition of *The Volunteer Monitor:* (graphs and figures not available)

Topic Areas

- **Macroinvertebrate Monitoring**

Tips

- **Tips for Statistical Analyses of Parallel Studies**
- **Watershed Activity Packet**
- **GREEN Public Workshops**
- **Speak Out! About Volunteer Monitoring**

Resources

- **IWLA Wetland Manual**

Survey

- **Survey for National Volunteer Monitoring Directory**

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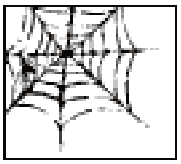

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From The *Editor*

Survey for New Directory

This map shows the locations of the 517 programs listed in the fourth National Directory of Volunteer Environmental Monitoring Programs, published in January 1994. What does the picture look like today? To find out, we need your help. Send in the survey form on page 23 by August 30 and put your program on the map! (and in the new Directory).

Next issue

The Fall 1997 issue of The Volunteer Monitor will focus on the theme of community outreach. The coediting group will be the Rivers Council of Minnesota.

Co-editing group for this issue: University of Rhode Island Watershed Watch, Kingston, RI

Graphic Designer: Brien Brennan

Printer: Howard Quinn Co., San Francisco, CA

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Monitoring Water Quality
U.S. Environmental Protection Agency Office of Water

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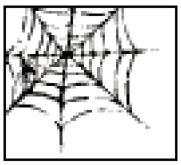

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LETTERS

Ohio Students Use "Stream Sentinel"

I was struck by an innovative technique presented in the Fall 1995 issue for using minnows to detect the presence of toxic discharges. The article described the Fort Worth Department of Environmental ManagementÕs homemade "stream sentinel," constructed from a plastic soft-drink bottle, which is stocked with minnows and placed in storm drain outfalls. (Note: see Fall 1995, p. 20, "Low-Cost Biological Monitoring.")

I've been working with students in Columbus, Ohio, for several years to assess conditions in their neighborhood stream, Adena Brook. The brook, like most urban streams, is highly affected by stormwater discharges. Though our chemical sampling p rogram has become quite sophisticated, it provides only intermittent glimpses of water quality. The Fort Worth idea impressed me as an inexpensive way to do around-the-clock monitoring.

A team of eight students from Dominion Middle School and Clintonville Academy was assembled in June of 1996 to try out the stream sentinel technique. With the help of the Columbus Division of Sewerage and Drainage, we selected three sites represen ting different storm drainage areas. Monitoring took place over an eight-week period in July and August and a four-week period in December. The students checked the fish every three or four days.

We found that the students had difficulty inserting the minnows into the small openings of the 2-liter soft drink containers. Also, we needed a more durable container that could withstand winter storms. We solved both problems by switching to 2- and 3-liter Ocean Spray juice containers, which have wide mouths and are made of heavy-duty plastic. For the floats we used 3/4" styrofoam boards with 3/4" plywood glued to the top. The plywood added strength, enabling the containers to weather high flow leve ls.

As the earlier article pointed out, the biggest headache is maintaining the fish. At the beginning of the project (summer) we bought minnows from a bait shop and took them to a pet store, which maintained them until we took them to the stream. Aft er school started, we tried raising the fish in the classroom. However, we had very high mortality in both the pet store and the classroom. For our December sampling, we bought minnows from the bait store and placed them immediately into the stream.

During the first few weeks of sampling, we had spotty survival of the fish at all our stream sites, possibly due to stresses associated with handling and with adapting to tanks at the pet store. By the second month (August) survival was greatly improved, and in December we had 100 percent survival at all sites.

In spite of our early difficulties with fish mortality, I feel this is an excellent method for volunteer groups. The students really enjoyed working on this project. Checking to see if their fish had survived was much more engaging than just going out and taking chemical measurements. They are now compiling their results for presentation at science fairs, and we plan to expand the project to an adjacent watershed by adding another school.

If readers would like more information about our project, my email address is jerry.wager@dnr.state.oh.us.

Jerry Wager
Ohio Department of Natural Resources
Division of Soil and Water Conservation
1939 Fountain Square Court
Columbus, OH 43224

Response: Thank you for your comments. I forwarded a copy of your letter to Brian Camp at Fort Worth Department of Environmental Management, who provided the following advice for using commercially raised minnows in the stream sentinel:

There are three scenarios that can lead to minnow mortality in a stream sentinel program: (1) Water pollutants are present in sufficient quantities to induce mortality; (2) The test minnows were weak due to stress or disease before placement in th e stream; and (3) The test minnows were mishandled during transport or introduction to the stream.

Scenario 1, of course, is what you are interested in-you are trying to detect pollutants in the stream. But if scenarios 2 or 3 are occurring, the fish may die even if the stream water quality is good-which invalidates your results. To minimize the possibility of scenarios 2 and 3, you need to make sure you are using healthy minnows.

Minnows bought from commercial sources such as minnow farms and bait shops are rarely suitable for immediate use in monitoring. Fish from these places tend to be stressed and diseased due to mishandling and overcrowding. Unfortunately, most people lack the resources to raise their own stock and are forced to purchase commercially grown minnows. If this is the case in your monitoring program, try to work out a way to quarantine the fish for at least 14 days before you use them in the stream sentine l. This is best accomplished in an aquarium because you can control all environmental factors. However, it can also be done by confining the minnows in a large, uncrowded minnow bucket in an unpolluted water body. Feed the quarantined minnows and remove d ead specimens daily.

Once you are sure you have healthy stock, treat the fish gently during any subsequent handling. Be very careful when netting and never let the minnows contact any type of dry surface. For transporting, fill a plastic bag to one-third capacity w ith water from the container holding the stock, add the fish, and seal the bag tightly with a rubber band, leaving a large air space above the water. If the fish will remain in the bag for more than about half an hour, inflate the bag fully with compresse d air before closing it (this can be done with an electric or battery-powered aquarium air pump).

Upon arrival at your monitoring site, take the temperature of the fish bag and the stream. Add 200 ml of stream water to the bag every 5 minutes until temperature equilibrium is reached. From this point on, add 50 ml of stream water every 30 to 60 seconds until the bag is full. The whole process should take at least 20 minutes. The fish should now be acclimated to stream conditions and ready for placement in the stream sentinel bottle.

With adverse stream conditions, you will need to allow even more time for acclimation, sometimes as much as one hour. By adding smaller amounts of stream water at longer intervals, we have successfully acclimated minnows to streams with pH read ings as low as 6 or as high as 10, and dissolved oxygen concentrations as low as 2.8 ppm.

Brian Camp
Fort Worth DEM
5000 Martin Luther King Fwy
Fort Worth, TX 76119
817/871-5458

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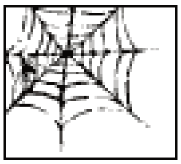	Note: This information is provided for reference purposes only. Although the information provided here was accurate and current when first created, it is now outdated.

Wanted!! Preferably Alive!

***by* Michael Beauchene**

I was voted class optimist the year I graduated high school, but the implications of this didn't hit me until about a year into my job as water resource biologist for Project SEARCH. My job description was simple-or so I thought. My duties: to develop and implement a macro- invertebrate monitoring program, using methods parallel to those currently used by the Connecticut Department of Environmental Protection (DEP). The resulting data would be utilized by the DEP for surface water assessment. The "catch": Project SEARCH is a high school moni-toring program open to all students regardless of scientific interest or ability.

Project SEARCH is a $1.5 million National Science Foundation"sponsored teacher enhancement grant administered through the Connecticut DEP and the Science Center of Connecticut. The project provides teachers with comprehensive training and equipment that enables them to successfully carry out water quality monitoring, wetland studies, and land use classification with their students. Over the past three years, SEARCH has trained teachers from 96 high schools, who in turn have brought over 2,500 students out to collect physical, chemical, and biological data.

The goal of the project is twofold: first, to provide high school students with a high quality, real-world, hands-on science experience, and second, to have students produce usable physical, chemical, and biological water quality data for local, state, and federal management officials.

Because of staff limitations at the Connecticut DEP, a real need exists for the students data. For example, the DEP has exactly one benthic biologist to cover the state's 5,830 miles of streams. The Department currently evaluates the macroinvertebrate community at 49 sites-most of them on major waterways with historical water quality problems-on a 6-year rotating schedule. Many small headwater streams are not assessed; the assumption made by the DEP is that these streams are a minor resource and are generally of high water quality. The student work provides the DEP with actual initial screening data. Without Project SEARCH, the water quality assessment at many of these small to mid-size streams would continue to be based on assumptions.

In order to meet both of SEARCH's goals (i.e., real-world science and usable data), the project's methodologies were based on those used by the DEP. For macroinvertebrate monitoring this meant the EPA's Rapid Bioassessment Protocol (RBP) III (Plafkin et al., 1989). We made just one modification: students identify the organisms to family level only, whereas DEP identifies to lower taxonomic levels. Twice in the fall and twice in the spring, students must perform all the following: evaluate the riffle habitat, collect and preserve a benthic sample, subsample a minimum of 100 organisms in the classroom, identify each organism to the family level, calculate six metrics (Taxa Richness, EPT Index, EPT/Chironomidae Ratio, Scraper to Collector-Filterer Ratio, Percentage of the Dominant Family, and the Hilsenhoff Biotic Index [HBI] family level), and compare their metrics to those of a DEP reference site. I told you I was an optimist!

Right off the bat (fall 1994), there were major problems with the student data. In tune with our goal of "real-world application," we had selected An Introduction to the Aquatic Insects of North America, by Merritt and Cummins, as the identification key for the students to use. After performing their identifications, the students placed preserved specimens in labeled vials and sent them to the project office for verification, in accordance with our quality assurance protocols. A major part of my job is to examine the specimens and verify the student identifications. Right away it was apparent that something was going terribly wrong. Only 12 out of 45 schools had been able to identify at least 100 organisms from their sample, and for those 12, the average correctly identified was 37% (range 20% to 60%).

This dismal data was a direct result of extreme frustration among students and teachers trying to wade through the terminology in the Merritt and Cummins key. Not only was the project failing to produce usable data, but even worse, there was no closure for the students. SEARCH was supposed to turn kids on to science-not away from it!

Sleepless night #357. I knew, and surveys of the teachers confirmed, that biomonitoring could be fun and exciting for the students. Teachers commented to me that the kids were amazed with the discovery of the macroinvertebrates. But all their enthusiasm was lost after reading a few couplets of Merritt and Cummins. Imagine you are back in the 10th grade and have the task of correctly identifying an aquatic insect nymph-or what appears to be an alien life form. Compounding your anxiety, you are using a key that requires you to make a series of choices between pairs like this: (1) "Terga and sterna of abdominal sections 1-9 separated by membranous pleural fold" or (2)"At most terga and sterna of abdominal sections 1-7 separated by membranous pleural fold." No problem, right?

The world of the average 10th-grade student simply does not include terminology such as "membranous pleural fold," "paraglossae," and "posterior supracoxal gills" -nor, probably, should it.

Looking for an alternative, I checked some macroinvertebrate keys produced by other volunteer monitoring groups (see "resources" box on page 5). These keys were easy to use and understand, but unfortunately most of their identifications went only to the level of order. For Project SEARCH, we needed to reach the family level.

To bridge the gap between volunteer keys and scientific keys, I decided to work with the students to create a family-level key, written in language they could relate to, for the organisms they were most likely to encounter. When I revisited the schools the following spring, I asked students to describe specific macroinvertebrate structures in their own words. Thus, for example, "tufts of ventral filamentous gills at the base of the coxa" was translated into "hairy armpits." Picture yourself in 10th grade once again. This time the key poses a question: "Does the organism have a tuft of fluffy gills at the base of each leg (hairy armpits)?"Yes or No."

Taxonomists around the world might shake their heads over such terminology-but when the students used the new key the next fall, the percentage of organisms correctly identified jumped to 70% (range 50% to 100%).

Unfortunately, it was out of the frying pan and into the fire. Now that identification was no longer a major concern, the students were struggling with calculating the metrics and analyzing their data. Many times the students reported moderately to severely impaired water quality at streams traditionally thought to have very high water quality.

Reviewing the students' data, I discovered a common scenario of limited diversity. The lack of diversity, which in most cases resulted from inadequate sampling procedures, lowered the bioassessment score, implying impaired water quality. But often the few families collected all had very low pollution tolerance values (HBI values of 0, 1, or 2). The student reports of "impaired conditions" were essentially an artifact of mindless number-crunching applied to a sub-optimum sample. A contributing problem was the inherent error in the family level HBI calculation. Since family tolerance values are based on all species in a family, a very sensitive species may have a score in the moderately sensitive range.

Thinking about these problems, I reflected that the EPA had published the RBPs as a guide for states, not high school students. Successful implementation of RBPs requires substantial training. Final water quality conclusions are based not on the metrics alone but on the combination of "best professional judgment" and the number-crunching. This is difficult if not impossible for students to accomplish on their first exposure to the subject.

Sleepless night #795. How could SEARCH ensure that high-quality streams were not being reported as low-quality streams? As I was racking my brain, I recalled an image from childhood: a Bugs Bunny cartoon in which Black Bart's image on a "wanted dead or alive poster" suddenly came to life. All at once an idea hit me. The next day I asked Guy Hoffman, the DEP's aquatic entomologist, to provide me with a list of organisms that met the following criteria:

- distributed across the entire study area (Connecticut)
- primarily riffle-dwelling
- sensitive to water quality impacts (Hilsenhoff tolerance value of 0, 1, or 2)
- readily identifiable by a distinctive structure or shape

I explained that the list should include organisms from several feeding groups (scrapers, shredders, collectors, etc.), and that it should not contain any organisms considered rare, threatened, or endangered.

Based on the candidates Hoffman provided, I selected six families and two genera for Connecticut's Most Wanted Macroinvertebrate List (see box next page), and created a series of Most Wanted posters complete with sketches, scientific names, "aliases," and descriptive information.

Connecticut's Most Wanted Macroinvertebrates

("aliases" are nicknames invented by SEARCH students)

- Pteronarcyidae, or giant stonefly
- Perlodidae, or perlodid stonefly
- Peltoperlidae, or roach-like stonefly
- Ephemerellidae, Drunella spp. (alias "body-building spiny crawler mayfly")
- Heptageniidae, Epeorus spp. (alias "2-tailed flat-headed mayfly")
- Glossosomatidae, or saddle-case- making caddisfly (alias "turtle shell caddisfly")
- Ryacophilidae (alias

In the fall of 1995, the Most Wanted posters were distributed to teachers along with the Project SEARCH teacher manual and student key. Students follow the same collection, preservation, and identification procedures as before, but now they are particularly on the lookout for the Most Wanted. When they capture a likely suspect, they check the poster for verification, then document the finding on their data sheet. The presence of Most Wanted organisms is also included in the school's summary report submitted annually to the DEP.

The Most Wanted list has been highly successful in accomplishing its major purpose-that is, to get the students away from "black box syndrome" in arriving at bio-assessments. The Most Wanted macroinvertebrates are all very sensitive to pollution, and since they all have a distinctive appearance students feel confident in their identifications. So if the metrics indicate moderate or severely impaired conditions at a site where a Most Wanted was found, an alarm is set off in the student's mind. Instead of blindly accepting the numbers, now the student must

"Michelin Man caddisfly")
- Blephariceridae, or net-winged midge (alias "Christmas tree fly")

use critical thinking: Did they collect an adequate sample? Did they identify all the bugs correctly?

Besides fulfilling this primary goal, the Most Wanted concept has produced a number of other benefits. For example, the Most Wanted list:

- Gives the DEP confidence in the students' data. Sites containing one or more Most Wanted macroinvertebrates have a high probability of having excellent water quality. (The probability increases greatly with the number of different types of Most Wanted found, and the abundance of each type.) Since students preserve the actual organisms and document when and where the sample was collected, a permanent water quality record is created. This allows the DEP to include student data in the state's 305(b) report to Congress. In 1994, student data added approximately 200 stream miles to the total reported as "assessed" in the 305(b) report.

- Gives students the satisfaction of seeing their data put to use.

- Provides students with a cross-check of their chemistry data. For example, if they recorded a pH of 1 but found a Most Wanted, they can wonder, "How could this be true?" and re-check their methodologies (did they calibrate the meter?).

- Reinforces the concept of an ecosystem. Students realize that physical, chemical, and biological parameters are interconnected, and that collecting data on just one reduces the strength of their conclusions about the water body.

- Helps SEARCH successfully fulfill both its objectives: to generate usable data, and to provide a real-world application of science for students in grades 9-12.

Does the Most Wanted list sound like something you would like to try? If so, contact your state aquatic entomologist(s) responsible for biological monitoring for help in developing your own list. Unfortunately, you cannot just "adopt" the SEARCH list (unless you are in Connecticut). Your list should and will be different; the taxa it contains will depend on your geographic location, the distribution of specific organisms in your area, and regional water quality standards. For additional information, please contact Michael Beauchene at the address below.

Michael Beauchene is Water Resource Biologist for Project SEARCH,
Office of Environmental Education,
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Hartford, CT 06106
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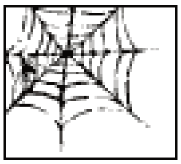

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Low-Cost Microscopes for "Bug" ID

Denise Stoeckel, a stream macroinvertebrate zoologist for the Illinois Natural History Survey and the quality assurance officer for Illinois RiverWatch, offers two suggestions for low-cost microscopes for macroinvertebrate identification. Both are mono cular scopes with a magnification of 20X, and both were designed as "student models."

The "MagiScope" is durable, portable, and easy for volunteers to use both in the field and in the lab. Stoeckel particularly recommends the model with the "Lumarod Illuminator"-a rod that draws ambient light to the subject, eliminating the need for mirrors, electricity, or batteries. Stoeckel notes that the field of view is somewhat small, but says this has not been a problem for volunteers. The MagiScope may be ordered from Nasco, 800/558-9595; cost is about $150 with the Lumarod, and $125 without.

An even less expensive scope that Stoeckel likes is the Wolfe EDS inclined monoscope. Stoeckel reports that it is durable and has a wider field of view than the MagiScope, but adds, "I don't know if I would recommend it for field use because it has no light source." The Wolfe EDS is available from Ward's, 800/962-2660, for about $80.

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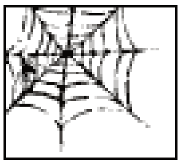

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Resources For Macroinvertebrate Monitoring

Project SEARCH Materials

The following three publications are available from Project SEARCH, Office of Environmental Education, CT DEP, 79 Elm St., Hartford, CT 06106; 860/424-3655.

Beauchene, M., L. Wahle, and S. Weiss. 1997. Water Quality Testing Manual for the Project Search Student Monitoring Program, 4th ed. 405 pages. Teacher's manual for Project SEARCH; covers methods for physical, chemical, and biological monitoring of streams and rivers. Each chapter is divided into teacher reference materials, teacher activity guides, and student materials. The manual includes Project SEARCH's "student-friendly" macroinvertebrate identification key and a set of posters for "Connecticut's Most Wanted Macroinvertebrates." $10.

Beauchene, M. 1996. "Macroinvertebrate Madness." 22 pages. An activity designed to help high school teachers introduce the concept of bioassessment. Students use hypothetical macroinvertebrate survey data to calculate metrics and determine level of impairment. No charge.

Beauchene, M. Project SEARCH 1995-1996 Annual Data Summary Report. 88 pages. Project description, QA/QC procedures and results, and student data. Free.

Resources designed for volunteers

Below are just a few of the excellent keys and manuals specifically geared toward volunteer monitors.

Division of Natural Areas and Preserves, Scenic Rivers Section. 1993. A Guide to Volunteer Stream Quality Monitoring. 23 pages. Outlines a user-friendly sampling protocol; includes key to orders (plus a few families), descriptions, and sample data sheets. Excellent for new programs. Developed for Ohio; useful in Midwest. Order from Ohio Dept. Natural Re-sources, 1889 Fountain Sq., Bldg. F, Columbus, OH 43224; 614/265-6453. Single copies free.

Fiske, S. and J. Byrne. 1988. Key to the Freshwater Macroinvertebrate Fauna of New England. 44 pages. Key to common New England taxa; includes several taxa usually not found in other keys. Available from River Watch Network, 153 State St., Montpelier, VT 05602; 802-223-3840. $5. A simple picture key is also available ($5); or order RWN's 250-page Benthic Macroinvertebrate Monitoring Manual ($25) and get both keys, plus detailed guidance for habitat assessment and macroinvertebrate monitoring.

Gilroy, M.P. and B. Furtado. 1995. Biomonitoring Guide. 150 pages. A comprehensive manual, aimed at high school students but applicable to adult groups as well. Includes dichotomous key to order level plus waterproof pocket-size key card. Order from Colorado River Watch Network, Lower Colorado River Authority, Austin, TX; 800/776-5272, x7634. $10. Also available: "Biological Activity Packet" ($8) with 12 classroom activities and "Biological Monitoring Video" ($10) demonstrating techniques for habitat assessment and macroinvertebrate collection and identification.

Hafele, R. and S. Hinton. 1996. Guide to Pacific Northwest Aquatic Invertebrates. 32 pages. A handy little field guide, printed on waterproof paper, that features color photographs of the 45 macroinvertebrates most commonly found in wadable streams of the Pacific Northwest. Each photo is accompanied by a brief description, including key identification characteristics. Order from Oregon Trout, 117 SW Front Ave., Portland, OR 97204; 503/222-9091. $9.95 + $1.50 S&H.

Illinois Department of Natural Resources. 1997. Illinois RiverWatch Stream Monitoring Manual. 92 pages. Specific to stream ecosystems of Illinois; contains a macroinvertebrate key, macroinvertebrate life cycle, and stream monitoring procedures for a variety of habitat types (riffles, leaf packs, snags, undercut banks, and sediments). Order from Illinois EcoWatch Network, c/o Nature of Illinois Foundation, 208 South LaSalle, Suite 2055, Chicago, IL 60604-1003; 312/201-0650. Free.

Kellogg, L.L. 1994. Monitor's Guide to Aquatic Macroinvertebrates. 60 pages. A pocket-sized guide including a key (with some important fly families), descriptions of major invertebrate groups, sampling protocols for both rocky bottoms and muddy bottoms, and sample data sheets. Excellent illustrations. Order from Save Our Streams, Izaak Walton League of America, 707 Conservation Lane, Gaithersburg, MD 20878; 800/BUG-IWLA. $5.

Scientific guides

Although the following standard references are quite technical and difficult for beginners, they nonetheless are essential for any serious macroinvertebrate monitoring program.

Hilsenhoff, W.L. 1988. Rapid Field Assessment of Organic Pollution with a Family-Level Biotic Index. J. N. Am. Benthol. Soc. 7:65-68.

McCafferty, W.P. 1983. Aquatic Entomology: The Fishermen's and Ecologists' Illustrated Guide to Insects and Their Relatives. Jones and Bartlett Publishers, Boston, MA.

Merritt, R.W. and K.W. Cummins. 1996. An Introduction to the Aquatic Insects of North America, 3rd ed. Kendall/Hunt Publishing Co., Dubuque, IA.

Plafkin, J. L., M. T. Barbour, K. D. Porter, S. K. Gross, and R. M. Hughes. 1989. Rapid Bioassessment Protocols for Use in Streams and Rivers: Benthic Macroinvertebrates and Fish. EPA/444/4-89-001. U.S. EPA, Assessment and Watershed Protection Division, Washington, DC. Available at no charge from Center for Environmental Research and Information, M.S. G72, 26 W. Martin Luther King Dr., Cincinnati, OH 45268-1072; 513/569-7562 (fax 513/569-7566).

Rosenberg, D.M. and V.H. Resh, eds. 1993. Freshwater Biomonitoring and Benthic Macroinvertebrates. Chapman and Hall, New York. 488 pages.

Attention RBP Users

The EPA is in the process of revising its Rapid Bioassessment Protocols. The draft version of the revised document is posted on the World Wide Web. To read the document, or comment on it, visit http://www.epa.gov/owow/monitoring/rbp/.

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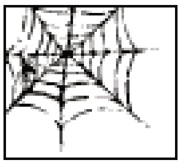

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The Care and Feeding of Dissolved Oxygen Kits

***by* Linda Green**

After 10 years as the University of Rhode Island (URI) Watershed Watch program coordinator, I tend to assume that I've seen every way to make mistakes. So imagine my surprise last fall when I opened a dissolved oxygen kit brought in by a volunteer for a reagent refill, pulled out two bottles that normally are filled with clear or pale pink reagents, and found both containing a dark brown liquid! Once again I was reminded of the importance of each and every ingredient in the kit, and of how things will go wrong even with what I thought was the best of attention.

The dissolved oxygen test is extremely popular among volunteer monitoring groups. In fact, the nationwide survey conducted in 1993 to gather information for the fourth edition of the National Directory of Volunteer Environmental Monitoring Programs found that dissolved oxygen was the third most common parameter measured, after water temperature and pH. There are three general ways to measure dissolved oxygen-performing the Winkler titration in a laboratory, using a dissolved oxygen meter, or using a dissolved oxygen kit (which is based on a modification of the Winkler titration). The reasonable cost, portability, reliability, and simplicity of kits make them the choice of many volunteer monitoring programs.

Over the years, URI Watershed Watch has come up with a number of practical tips and guidelines for getting top-notch results from our dissolved oxygen kits. (The pointers in this article are particularly applicable to LaMotte Company dissolved oxygen kits, since that is the type we use.)

Storage

Don't store your dissolved oxygen kit (or any kit, for that matter) in the trunk of your car. First of all, all that bouncing is not a good idea. Secondly, extreme summer or winter temperatures in the trunk will hasten the degradation of the chemicals. Storing a kit on a sunny shelf in your home or office will have the same effect. The cabinet under your kitchen sink is not a good choice either, because it tends to be humid. And for safety's sake, be sure to keep any monitoring kit out of the reach of exploring children. A shelf in a dry closet without temperature extremes is a good place for storing the kit.

Protecting reagents

How do you know that the reagents in your dissolved kit are in good condition? First, take a look at them! All the liquids should be clear, without cloudiness and without particles floating in them.

The most critical reagent is the titrant, sodium thiosulfate. Any change in the concentration of the titrant will directly affect the results of the test. According to LaMotte Technical Service Representative Steve Wildberger, it is essential to replace the sodium thiosulfate each year. It's also important to guard against contaminating this reagent. If you refill the syringe between samples, wipe off the tip before putting it back in the sodium thiosulfate bottle. This helps prevent Òsample splashÓ from getting into the reagent bottle. The sample now contains iodine (if it's yellow), which will neutralize the titrant. If there's chlorine in the water, this will also reduce the strength of the titrant since chlorine reacts with sodium thiosulfate. Finally, if you have leftover titrant in the syringe after testing all the samples, resist the temptation to be thrifty-DO NOT put the leftover titrant back into the reagent bottle. Discard it along with the sample and other wastes. In fact, a good basic rule of thumb for any reagent is "once it's out of the bottle, don't put it back in."

Although some monitors wrap the individual reagent bottles with tape or aluminum foil to further exclude light, Wildberger does not recommend this. He points out that light-sensitive reagents are supplied in amber bottles to protect them from light, and that covering the bottles with foil or tape makes it difficult for monitors to inspect the contents.

Cleaning glassware

Each time you finish using the kit, rinse out the sample bottle and titration vial with tap water. Detergent is both unnecessary and inadvisable. (Note: We usually use the term "titration vial" for what LaMotte calls the "titration tube." Since it looks like a vial, our monitors find the term "tube" confusing.)

You don't have to rinse the titrator syringe between uses. However, remember to store the syringe with the black neoprene tip backed off from the bottom. This helps prevent the tip from getting stuck. Another way to prevent sticking is to coat the tip with a tiny bit of silicone grease, available at hardware or plumbing stores. (Be sure to use solid greaseÑnot spray, which will destroy the neoprene.)

After rinsing, make sure all items have dried completely before you put them back in the kit. You don't want damp bottles to turn your kit into a humidor, especially if you are using powdered reagents such as sulfamic acid. (Some LaMotte kits contain sulfamic acid powder instead of sulfuric acid.)

In time, the titration vial may acquire a blue tinge. If this occurs, use a bottle brush to gently scrub off the discoloration. Don't use bleach, because residual bleach could cause a falsely elevated result next time you analyze a sample.

Refilling reagent bottles

If a volunteer needs a refill during the monitoring season, we discard any reagent remaining in the bottle, rinse the bottle with a little fresh reagent (discarding the rinse reagent), and fill the reagent bottle. Each fall, after sampling season ends, we ask all volunteers to turn in their kits for a complete overhaul. We inspect the kits and then empty, clean, and dry all the bottles. The following April, just before the start of the next season, we fill all the bottles with brand-new reagents from bulk reagent refills. We order the bulk refills shortly before we need to use them, as LaMotte recommends, and we never save them from year to year.

Checking volunteers' technique

Twice during the six-month sampling season, we require our volunteers to bring their kits to our laboratory for a quality control check. We provide two tubs of water for dissolved oxygen testing. One tub has an elevated dissolved oxygen content (approximately 8 ppm), which we achieve by bubbling air through the water with an aquarium aerator. In the other tub, we lower the dissolved oxygen level to about 2 ppm by bubbling nitrogen (purchased from a gas supply company) through the water. (Note: The gases need to bubble through the water for at least half an hour before the first sample is taken, and must be bubbled through continuously until all samples have been taken.) Volunteers use their kits to test two water samples from each tub; their results are then compared to results obtained by program staff using their own kits. By the way, one thing I've learned is to keep my mouth shut while the volunteers are working. The distraction of keeping up a conversation can cause them to make mistakes.

While our volunteers are running their tests, we're keeping an eye on their technique. (We find that volunteers welcome this attention; they want to make sure they are performing the test correctly, and that their time and effort are not being wasted.) The most glaring error is when volunteers pour off a little water from the top of their sample bottles, in the mistaken belief that they need to make space to add the reagents. The best antidote for this practice is a quick demonstration in which you run two samples side by side, one with the bottle full to the brim and one with "just a little water poured off." Actually seeing the higher dissolved oxygen content of the bottle with the air space is a powerful educational tool! We also point out that the 21% oxygen content of air is equivalent to 210,000 ppm oxygen, which is why even a small air bubble can significantly affect the dissolved oxygen measurement.

So, what was the cause of the brown gunk in those two reagent bottles? Probably I'll never know for sure; my guess is that the bottles were somehow switched when the reagents were refilled. I have, however, kept those two bottles on my desk as a reminder to stay vigilant-and expect the unexpected.

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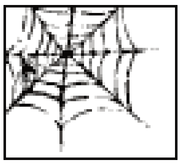

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Common Questions About DO Testing

Q. Should I pour off any of the water in my sample bottle before I add the reagents?
NO! Pouring off some of the water allows space for an air bubble to be trapped when the bottle is capped. When you shake the bottle this oxygen mixes with the sample and causes erroneously high results. It's OK for some liquid to overflow as you add the fixing reagents. (If you are concerned about spillage, put the bottle on a paper towel.)

Q. How should I hold the dropper bottles to dispense the reagents?
A. Hold the dropper bottles completely upside down (i.e., vertical). This ensures a uniform drop size.

Q. Sometimes after I add the sulfuric acid some brown "dots" remain. Is this OK?
The brown particles should be dissolved before you continue the test. Try shaking the sample bottle again. If this doesn't work, add one more drop of sulfuric acid. (Occasionally in water with an algal bloom there may be some organic matter that won't dissolve. However, you should be able to tell the difference between this and the chemical particles.)

Q. What is meant by saying that the sample is"fixed"?
After the sample is fixed (i.e., after the first three reagents are added), contact with atmospheric oxygen will no longer affect the test result. Fixed samples may be stored up to eight hours, if kept refrigerated and in the dark.

Q. When filling the syringe with the thiosulfate reagent, how far back should I pull

the barrel?
The point of the black neoprene tip should be set right at zero. This is extremely important.

Q. Exactly how should I read the syringe?
Hold the syringe vertically with the tip pointing down. Read the point where the black tip of the plunger meets the scale. The bigger lines represent ppm (1 ppm, 2 ppm, etc.) and the smaller divisions represent two-tenths (0.2 ppm, 0.4 ppm, etc.).

Q. OK, now I'm titrating. How come the drops of sodium thiosulfate seem to be running down the side of the vial, instead of dropping directly into the sample?
The cap of the titrator vial should have a tiny vent hole that allows displaced air to escape as sodium thiosulfate is added. If the vent hole is absent or too small, or if it has gotten covered by a drop of liquid on top of the cap, the sodium thiosulfate will run down the side of the bottle instead of dropping into the sample. You should first check to be sure the vent hole is unobstructed. If necessary, you can create (or enlarge) a vent hole by heating a pin and pushing it through the plastic.

Q. The directions say to add sodium thiosulfate until the sample turns a straw yellow, then add the starch indicator. Why shouldn't I add the starch indicator all at once in the beginning?
If your sample is already pale yellow right after it is fixed, you should go ahead and add the starch indicator before you begin titrating. But if it is brownish, you should titrate it to a pale yellow before adding the starch indicator. LaMotte Technical Service Representative Steve Wildberger explains that you risk overshooting the endpoint if you add the starch indicator at the beginning, because the change from blue to colorless is much more abrupt than the gradual change from brown to yellow. He also notes that the color you're looking for is more of a"manila folder yellow" than a"straw yellow." Some other pointers for accurate titration:

- Hold the titration vial against a white background.
- Swirl the sample after each drop of sodium thiosulfate is added.

Q. What if my sample is colorless after it's fixed?
This means there is no dissolved oxygen in the sample. If this happens, you might want to test a sample that you know contains oxygen to make sure that your kit is functioning properly. (One way to do this is to intentionally introduce an air bubble into the water sample, shake well, then fix the sample. You should see a yellow color.)

-Linda Green

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Making a Standard Solution for Dissolved Oxygen

by *Mark Mattson* and *Peter Kerr*

In the accompanying article on the "Care and Feeding of Dissolved Oxygen Kits," Linda Green describes how URIWW volunteers come to the program lab for a quality control (QC) check of their dissolved oxygen kits and technique. But it's not always possible for volunteers to come to a lab for QC testing. At Massachusetts Water Watch Partnership (MassWWP) we use a different approach for QC of dissolved oxygen kits: we prepare a biiodate standard solution1 which we mail to our volunteers.

While it's relatively simple to make a standard solution for chemicals like phosphate or nitrate, making a standard solution for dissolved oxygen is more difficult because the concentration of dissolved oxygen in water changes as soon as the solution is exposed to air. To get around this problem, the standard contains biiodate as a "stand-in" for oxygen.

The biiodate standard solution is stable at room temperature for 6 months or more and can be sent through the mail if tightly sealed, bagged, and boxed. We dispense the standard into brown polyethylene 500-ml bottles, which we mail to the volunteers without telling them what the expected value is. The volunteers test the QC standard using their dissolved oxygen field kits, then report their results back to us. We set a limit of 10% error; if this is not met, the thiosulfate reagent must be replaced.

Preparing the standard

(Note: These instructions are for a 20-L batch. At MassWWP we make a large batch so we can send some to all our volunteers. However, the recipe can be scaled down if you want to make a smaller amount.)

1. Measure 20 L of distilled water into a large container, or carboy. Mark the carboy at the 20-L point (be sure to place the mark at the meniscus of the liquid). Then remove approximately 1 L of water (the amount removed is not critical; you are just making room in the carboy to add other ingredients).
2. Dry potassium biiodate, KH(IO3)2, for 2 hours at 103-105°C (once dried, it can be stored in a desiccator for future use). Carefully weigh out an amount between 300 and 800 mg. The amount of biiodate used will determine the equivalent "dissolved oxygen" content of the standard (see below for formula to convert mg/L biiodate to equivalent mg/L dissolved oxygen). Record the exact weight, to the nearest mg. Dissolve the biiodate, with stirring, in a beaker with about 500 ml distilled water. You may need to crush the biiodate crystals with a stirring rod to help them dissolve.
3. To the 19 L of distilled water in the carboy, add 20 ml of 5N NaOH and approximately 200 g potassium iodide, KI. Mix with a stir bar to dissolve. Then add the 500 ml biiodate solution to the carboy and mix. Finally, add distilled water to bring the level in the carboy up to the 20-L mark.

    [1] The potassium biiodate standard for the Winkler dissolved oxygen test (Method 360.2) is described in U.S. EPA, Methods for Chemical Analysis of Water and Waste, EPA-600/4-79-020.
4. The equivalent dissolved oxygen (DO) concentration of the solution is calculated with the following formula:

    mg biiodate/L x 0.2462 = mg DO/L

For example, if you added 660 mg biiodate to 20 L, the solution contains 33.0 mg biiodate/L, so:

33.0 mg biiodate/L x 0.2462 = 8.12 mg DO/L

Before sending the DO standard out to our volunteers, we always test it using the same type of kit the volunteers will use. We then compare the volunteers' results to the results we actually obtained (not to the theoretical DO concentration calculated with the formula).

Using the standard for QC

Important: Be sure to emphasize to your volunteers that the procedure for testing the dissolved oxygen standard is different from the procedure they usually follow for water samples. The main difference is that they do not add the first two reagents (manganous sulfate and alkaline potassium iodide azide, or powder pillows #1 and #2 in the Hach kit).

To use the QC standard, measure approximately 60 ml of the standard into a beaker and add the sulfuric acid reagent (or Hach powder pillow #3). Mix for 3 to 5 minutes. The solution should turn yellow, indicating the formation of iodine. Treat this as a "fixed" water sample and follow your usual procedure-that is, carefully measure the sample volume you usually use for titration (this volume will vary depending on type of kit), then titrate with thiosulfate.

How the biiodate standard works

Dissolved oxygen test kits are based on the Winkler titration method. In this method, reagents are added to the water sample to produce a series of chemical reactions whose net result is the production of iodine in direct proportion to the amount of dissolved oxygen originally present in the sample. Titration with thiosulfate is then used to determine the concentration of iodine.

The QC standard contains biiodate, KH(IO3)2, which is converted to iodine when the acid reagent is added. Since you know how much biiodate you used to make the QC sample, you know how much thiosulfate should be needed to titrate the iodine. If you use more or less thiosulfate than expected, there is probably something wrong with the thiosulfate reagent.

Note that the biiodate QC standard only checks the performance of the thiosulfate. It doesn't tell you whether the manganous sulfate or alkaline potassium iodide azide solutions are performing properly. However, the thiosulfate reagent is the most critical one for this test. Mark Mattson is a Senior Scientist and Peter Kerr is Laboratory Supervisor at the Water Resources Research Center, Blaisdell House, University of Massachusetts, Amherst, MA 01003-0820; 413/545-2842. Both are also technical consultants to MassWWP.

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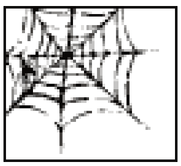

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Guarding Against Reagent Degradation

by Tom McAninch

The reagents in test kits are chemically reactive; otherwise they would not be useful for quick and easy field tests. Because they are reactive, test reagents can easily degrade. Test reagents are also very susceptible to contamination under field conditions.

Both degradation and contamination can cause readings to be in error, invalidating many hours of monitoring effort. So, how do you prevent these problems-and, if they do occur, how can you recognize and correct them?

Recognizing bad reagents

One of the easiest ways to tell if a reagent has gone bad is by its appearance. When a reagent is received, note its appearance. Each time you use it, look for any change in color or clarity, or for formation of solids (e.g., a crust on the lip of the bottle, floating particles, or solids settled at the bottom). Also check expiration dates, and replace reagents before they expire.

If you suspect that a reagent has gone bad, you can test it by using a solution of known concentration. Instructions for making standard solutions may be found in Standard Methods.[1] Alternatively, you can purchase standards from kit manufacturers or other chemical companies, or obtain them from labs you work with.

You can also test your reagents by checking the suspect kit against another kit which has fresh reagents, or by cross-checking the kit against another method, such as a meter.

Remove and discard any bad or expired reagents (check the Material Safety Data Sheet, included in kits, for safe disposal methods), and restock the kit with fresh reagents. Temperature decomposition

In general, the speed of a chemical reaction doubles with every 10°C increase in temperature. Since reagent decomposition occurs by means of chemical reactions, high temperatures will speed decomposition. Thus, a kit that is routinely stored in a car trunk or in direct sunlight can be expected to decompose three to four times faster than a kit protected from high temperatures.

Prevention:

- Store kits away from heat and sunlight.
- During testing, do not expose reagents to heat or sunlight any more than necessary.

Photochemical decomposition

Many reagents will photochemically decompose when exposed to direct sunlight. Reagents containing silver are especially sensitive to sunlight, and will turn black when photodecomposition occurs. Kits that use a titration method to test chloride or salinity generally include a silver nitrate titrant. This reagent is supplied in an amber glass, amber plastic, or other opaque plastic container to protect it from sunlight. Some volunteer monitoring groups wrap the bottles in electrical tape or foil to further exclude light, but this is not necessary-in fact, it's undesirable because it makes it difficult to visually examine reagents for signs of degradation.

Prevention:

- Store kits away from light.
- Keep reagent bottles inside kit during storage.
- When performing tests, do not leave reagent containers open any longer than necessary.
- When replacing or refilling sunlight- sensitive reagents, be sure to place them in opaque bottles.

Air decomposition

Air is the enemy of several reagents. Evaporation can concentrate reagents, which is especially critical for a titrant. Also, many reagents can react chemically with oxygen or carbon dioxide in the air. For example, the manganese reagent used in the dissolved oxygen test reacts with oxygen.

Prevention:

- When performing tests, do not leave reagent containers open any longer than necessary.
- Keep reagent bottles tightly capped.

Contamination

Contamination is probably the biggest enemy of monitoring kits. Any foreign material introduced into a reagent bottle can cause test results to be too high or too low. If mold or algae get into starch reagents, they can grow there since starch is a nutrient.

One common source of contamination is inadequate cleaning of equipment that is used to analyze more than one sample. Another is failure to use a dedicated dropper for each reagent. To avoid dropper-related contamination problems, some kits use bottles with built-in dropper tips (the bottle is inverted and squeezed to expel drops of reagent). Such bottles eliminate the danger of putting a dropper into the wrong reagent bottle, and also protect reagents from air and contaminants.

Prevention:

- Use dedicated droppers and dedicated equipment wherever possible.
- Always bring a container to the field for washing equipment between tests.

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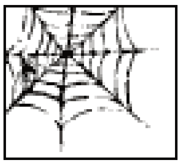

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Liquid Versus Powdered Reagents

If you're planning to use a test kit, you may be wondering whether to choose one that provides reagents in liquid form or as a powder (generally in individual packets or "powder pillows"). Here are a few pros and cons to consider. People who prefer powdered reagents say they consider them safer than bottled reagents, which can spill onto the user's skin. They also point out that powdered reagents are more stable and that an individual powder pillow is less vulnerable to contamination than a bottle of liquid. People who prefer liquid reagents say they are easier to use, noting that powder packets can be hard to open, that powder can spill or be blown by the wind during pouring, that it's sometimes hard to get all the powder out of the packet, and that some powdered reagents are hard to dissolve whereas liquids go into solution readily and completely.

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Fourth Secchi Dip-In

Over 2,000 volunteers are expected to participate in this year's Great American Secchi Dip-In, June 27-July 13. Like its three predecessors, the Dip-In will provide a snapshot of transparency in lakes, estuaries, and rivers throughout North America. It's also a good opportunity for raising public awareness about water quality.

Maps based on previous Dip-Ins have shown that the northern parts of the U.S. and Canada have the clearest lakes, and that lakes in agricultural regions have some of the lowest transparencies. The deepest transparency found so far was 52 feet, in a Minnesota lake. The Dip-In is sponsored by the North American Lake Management Society and the U.S. EPA, and directed by scientists at Kent State University. For more information, visit the Dip-In Web site at http://humboldt.kent.edu/~dipin, or contact Dr. Robert Carlson, Dept. Biological Sciences, Kent State University, Kent, OH 44242; 330/672-3849; email RCarlson@Kent.Edu.

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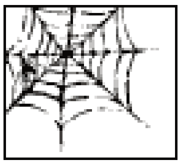

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Disposal of Chemical Wastes

by Steve Wildberger

Any monitoring program that performs chemical testing faces the problem of disposing of chemical wastes. These include both individual unused reagents and "reacted sample waste" (waste generated during performance of the tests).

Unused reagents

Disposing of expired or "extra" unused reagents is usually the monitoring program coordinator's job.

Tip: Program coordinators can reduce or eliminate the problem of leftover reagents by ordering quantities that take into account the reagent's shelf life and the program's rate of usage. Instructions for disposal of individual reagents must be included on the Material Safety Data Sheet (MSDS) provided by the manufacturer. Modest quantities of many common test kit reagents can be safely diluted and disposed of down the drain if precautions regarding incompatibility are observed. For example, acids and bases should be neutralized before disposal (see instructions on MSDS). Note that neutralization often occurs as part of the test procedure, making the disposal of reacted sample waste easier than reagent disposal. For example, the reagents used in the dissolved oxygen test include both an acid (sulfuric acid) and a base (alkaline potassium iodide azide), but these neutralize each other when the test is performed.

Often the final instruction on the MSDS is "dispose of in accordance with applicable federal, state, and local regulations." These regulations often include various exemptions which relate more to the classification of the waste producer (i.e., households vs. industry) than to the chemicals involved. Monitors need to become aware of the regulations for their area (bearing in mind that local jurisdictions, such as counties, can set their own guidelines) and make a plan that follows the regulations and emphasizes prudent and responsible practices.

Tip: Form an alliance with the laboratory personnel at the agencies that deal with waste disposal regulations for your area. They should be familiar with the tests you are performing and can provide advice and support. It's also very helpful to contact local government recycling coordinators and find out about household hazardous waste programs.

Reacted sample waste

The challenge of disposing of wastes from reacted samples generally falls to individual monitors. Monitors should be sure to bring a designated waste receptacle to their site whenever they perform chemical testing.

The majority of chemical tests commonly used in volunteer programs produce wastes that can be safely disposed of down the drain if flushed with plenty of water. These include field kits for testing dissolved oxygen, pH, phosphorus, turbidity, and alkalinity. See the box below for guidelines from GREEN on disposing of non-hazardous sample wastes.

A few test procedures include heavy metals that are not neutralized in the test procedure. Those most commonly used by volunteer groups are the cadmium reduction method for nitrate, the Nessler method for ammonia (which uses mercuric chloride), and the argentometric or mercuric nitrate methods for chloride or salinity.

Tip: Be sure to keep wastes that contain heavy metals separate from wastes that can be safely disposed of down the drain.

If your program is partnered with a lab, the lab can separate the heavy metals from the rest of the sample waste by precipitating the metal and filtering it out. Alternatively, the waste can simply be evaporated (in a safe place, out of reach of children) to reduce the volume of waste that needs special handling. The solid residue should be disposed of as you would Ni-Cd batteries or other heavy metal waste (usually in a landfill).

Alternate methods

Efforts should be made to incorporate alternative methods that reduce or eliminate the use of toxic materials. However, alternative methods should be carefully evaluated for "hidden" hazardous materials. For example, in the Spring 1993 issue of The Volunteer Monitor, Robert Frease compares the relative hazards of salinity titration kits versus hydrometers in his program. While hydrometers are theoretically less toxic, Frease believes that "more potential toxicity is associated with the disposal of broken hydrometers." Frease notes that the hydrometer counterweight contains the heavy metals lead, tin, and antimony, and points out that the program annually disposes of less than 1/5 pound of silver from salinity kits.

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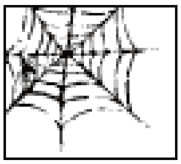

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GREEN Guidelines for School-Based Programs

Thousands of students and teachers throughout the world participate in GREEN (Global Rivers Environmental Education Network). Most of them perform the water quality tests described in Mitchell and Stapp's Field Manual for Water Quality Monitoring,1 including field tests for five chemical parameters: dissolved oxygen (DO), biochemical oxygen demand (BOD), pH, total phosphorus, and nitrate. GREEN provides teachers with the following guidelines for disposing of the waste generated from field test kits.

Two types of waste are generated during the chemical analyses: those classified as non-hazardous (to both humans and the environment) and those classified as hazardous. These two types of waste must be separated for proper disposal. All wastes from the DO, BOD, pH, and total phosphorus tests are classified as non-hazardous. If you are using a nitrate kit that employs the cadmium reduction method, the waste from this test should be treated as hazardous waste due to the cadmium present in solution.

Disposing of non-hazardous wastes There are two options for disposing of the non-hazardous wastes:

1. For disposal in a sink: Bring an empty plastic reagent bottle, or a glass container (such as a Mason jar), to your site and use it to collect all the wastes from the DO, BOD, pH, and total phosphorus tests. These wastes can safely be mixed together with no possibility of explosions or forming toxic gases. Take the container of

waste back to the lab and dispose of the waste in the sink. Be sure to run plenty of water through the sink in order to prevent corrosion of the pipes.

2. For disposal in a trash receptacle: Collect the wastes in a container (an old coffee can works well) filled with an absorbent material like kitty litter. The kitty litter will absorb the water and bind up the waste. This container should be tightly covered and may be placed in a trash receptacle as long as there is no free-flowing liquid remaining.

Disposing of hazardous wastes

If your kit uses the cadmium reduction method for nitrate, the wastes generated contain small amounts of cadmium. This metal is classified as a hazardous waste by the Environmental Protection Agency. Wastes from the nitrate test should be collected separately in a container that is clearly marked (e.g., "toxic waste jar"). This waste can be concentrated by slow evaporation under an exhaust hood. Check with the chemistry teacher or science department head at your school for the proper procedure for disposing of this waste.

[1] Available from GREEN, 206 S. Fifth Ave., Suite 150, Ann Arbor, MI 48104; 313/761-8142.

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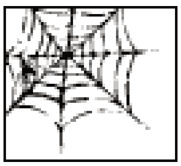

Nutrients Test Kits: What Can We Expect?

by Revital Katznelson

Suppose you are monitoring an urban creek on a limited budget and you want to run tests for nutrients (phosphate, ammonia, nitrate). You intend to use inexpensive field kits-the kind that rely on a visual color comparison, using a "color wheel" or "color comparator." How would you go about shopping for the most suitable kits? I look for reagents that produce a blue or green color (e.g., salicylate method for ammonia) rather than yellow (e.g., Nessler for ammonia), because the human eye is better at perceiving density of blue or green. I look for less toxic reagents (e.g., salicylate versus Nessler for ammonia, zinc rather than cadmium for nitrate). I look for the lowest range, relying on the option of diluting my sample if the concentration is too high (and then make sure I have the equipment for making dilutions-a small syringe without needle, distilled water, and a dedicated jar). I look for reagents in liquid form rather than powder because I find it tedious to wrestle with those packets and with the wind that blows the powders all over the place.

You would be amazed at the colors you get when you test distilled water with some of these kits. It's as if the reagents have a life of their own. So, the first thing to do with a new kit is to run a reagent "blank," using distilled or deionized water (supermarket grade will do) in place of the sample. In my work with volunteer monitoring groups in Alameda County, we found that for the ammonia kit the blank read as 0.1 ppm (parts per million). For that kit we defined our "reporting limit" as 0.2 ppm, because that was the next increment of color we could be certain was higher than the blank. Anything below this value is uncertain, so when we read color in the range of 0 to 0.2 ppm with our creek sample we normally report the result as "<0.2 ppm."

Many volunteer monitors, including many classroom teachers, currently use inexpensive field kits to test for nutrients. If they are testing in relatively unpolluted waters, they may be misled or confused by the results. Sometimes kit users see a value of zero and think there are absolutely no nutrients in the water body so nothing should be able to grow, yet they see algae growing in abundance. So, how about replacing the concept of "zero" with the concept of "below reporting limit"? Then you can visualize just a few ions of ammonia, nitrate, or phosphate in the water, and you know you cannot detect them with your kit. Remember: a nutrient has to be depleted to extremely low levels before it becomes a "limiting factor" for growth, and some algae and microorganisms have invented amazing adaptations to scoop up nutrients even if they are present in minute amounts. In fact, some of us ecologists can tell you exactly which nutrient is in low supply based on the species of algae we see in a lake.

So, why use these kits anyway? Well, for one thing, they are very affordable. The more sensitive methods for measuring nutrients require the use of a colorimeter ($250 and up) or spectrophotometer (over $1,000) to read the color change, and often require lab facilities as well. And the kits do provide valuable information in situations where nutrient levels are relatively high. Kits are extremely useful in tracking the sources of nutrients further up the watershed-for example, in determining which outfall brings the most nutrients into the creek during a storm event. They can also be useful in characterizing spatial trends (e.g., do we have more nitrate upstream or downstream?) or temporal trends (are the phosphate concentrations in July higher than in January?). And they can alert us to serious pollution problems. For example, a kit can tell us whether fish are threatened by ammonia toxicity. Why do we need to know how precise (reproducible) and accurate (close to the "true" value) our measurements are? How can we decide what range of error we can tolerate for each nutrient and for each range of concentrations? This depends on the ecological significance of each variable at each range of concentrations. For example, the difference between 2 and 4 ppm phosphate, or 0.2 and 0.4 ppm ammonia, will not have a profound effect on creek critters, so we can tolerate an error of 50% or even 100% in these cases. But the difference between 2 and 4 ppm ammonia (atpH of 8.5 or higher, because ammonia is toxic at higher pH) will mean life or death to some fish, so we want to make sure that our readings are reliable within 5% or 10% error.

It is actually quite simple to adapt the formal laboratory methods of evaluating precision and accuracy and use them with nutrient field kits. It is also necessary, especially where accuracy is of concern. Use of standards is highly recommended; however, I have obtained inconsistent results with some kits. For example, a nitrate standard of 5 ppm was read as 1 ppm by a kit. The problem is that even with the best intentions and the best research and development and the best plastic and pigment chemistry, kit manufacturers cannot produce an absolutely, universally accurate kit for any analyte. It is the responsibility of the user to assure the reliability of data obtained with kits, and a protocol telling the user how to do that needs to be written.

I am currently in the process of examining a number of questions related to kits, including limitations of kits and issues of data quality assurance. In a future issue of this newsletter, I hope to be able to provide some answers, as well as some specifc advice based on real problems encountered when using these kits in urban creeks. Meanwhile, for volunteer monitors who are using nutrient kits and who want to generate reliable data so that their efforts "count," my advice is: don't use the kits without training, always maintain access to technical support and ask questions, and never agree to use the kits if you do not also agree to use your brain, your eyes, and your common sense (and maybe a little math).

Revital Katznelson is a water specialist with Woodward Clyde Consultants and is working on incorporating nutrient monitoring into volunteer monitoring programs in Alameda Co., CA. She can be reached at 510/874-3048.

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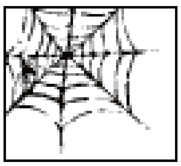

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Salinity by Conductivity and Hydrometer

A METHOD COMPARISON

by Peter Bergstrom

Why measure salinity?

Salinity is critically important to understanding estuaries (bodies of water in which fresh and salt water mix-such as bays, tidal rivers, or salt marshes). Salinity measurements are used to study estuarine circulation and assess the effects of freshwater flow and tides. In particular, salinity can help reveal whether development is affecting the quantity of freshwater input to the estuary. Salinity also defines suitable habitats for plants and animals, so measuring salinity is important when conducting surveys or planning restoration programs.

Salinity testing methods

For any or all of the above reasons, volunteer monitoring groups often decide to measure salinity. And then things can start to get interesting, because there are four major methods to choose from-chloride titration, hydrometer, refractometer, and conductivity. (All four were discussed by Steve Wildberger in The Volunteer Monitor, Spring 1993.) None is perfect; each has its pluses and minuses. But wait-it gets even more complicated. Each of the four methods measures something different: chloride titration measures chloride ions, a hydrometer measures specific gravity, a refractometer measures light refraction, and a conductivity meter measures electrical conductivity. In each case, the factor that is actually measured is converted to salinity by means of some type of formula. Because they measure different things, each method may give a slightly different result for the same sample. This can become a problem if you want to combine or compare your salinity results with data that were collected by another method.

Only conductivity can be measured using an electronic meter, and that makes it the method of choice for almost all professional water quality monitoring. (See box on next page for more on conductivity meters.) This does not mean that conductivity is an inherently better measure of salinity. However, it does mean that volunteer monitors-relatively few of whom use conductivity meters-may need to address the issue of method differences if they want to compare their data with agency-collected conductivity data.

How can you find out how closely your salinity measurements match conductivity data? And if you find a discrepancy, how can you adjust your data to make it more compatible? When Magothy River Association (MRA) volunteer monitors wanted their hydrometer salinity data to be used together with their state agency s data, they investigated these questions with a method comparison study.

Magothy River volunteer monitoring

The Magothy River is a tidal tributary of Chesapeake Bay, located in Maryland just south of Baltimore. It is fairly short (about 7 miles to the upstream limit of tidal influence) and most of the tidal portion is mesohaline, with a mean surface salinity between 5 and 18 parts per thousand (ppt). Submerged aquatic vegetation (SAV) in the Magothy declined dramatically in the 1970s and has only recently started to increase. In response to the SAV decline, the MRA volunteer monitoring program was founded in 1983. At first, the volunteers measured Secchi depth and water color. They started measuring salinity (using hydrometers) in 1987, and the program has since added a variety of other physical and chemical parameters.

In 1991, I joined the program and began sampling in two creeks. The following year I expanded my efforts, teaming up with several other MRA volunteers to sample-by boat-12 to 18 stations in the Magothy mainstem. I also continued my creek sampling.

At first we used the same methods as the other MRA volunteer monitors, including hydrometers for salinity and a kit for pH. Then, in 1993, I tried using pocket conductivity and pH meters-mainly to save time, since we were sampling many stations in rapid succession. Recognizing that there might be differences between the old and new methods, I began collecting comparison data, using both methods on samples from the same bucket. I found that the pH meter results (Oakton pHTestr 2) agreed closely with the results from the old method (LaMotte wide-range pH kit #2117), so I discontinued comparison testing for pH. However, the 1993 salinity data did show some method differences, so I continued collecting salinity comparison data through 1996.

Hydrometer versus conductivity

By the end of 1996, I had 546 pairs of salinity comparison data (459 on the mainstem and 87 on the creeks)-more than enough to do a statistical comparison between the two methods. I collected the data between April and October, over a salinity range (by hydrometer) of 1.5 15 ppt. The data included three fairly wet years and one dry year, which extended the salinity range.

For measuring conductivity I used a temperature-corrected pocket meter (Oakton TDSTestr4, range up to 19.9 mS/cm) in 1993 and 1994, and a Hydrolab Surveyor III meter (provided by the U.S. Fish & Wildlife Service) in 1995 and 1996. Before recording conductivity data, I waited for the thermistor used to measure temperature in the meters to stabilize (which can take 3 5 minutes). I measured specific gravity with a LaMotte hydrometer, and calculated salinity with an equation based on revised tables published by LaMotte in 1993 (see The Problem with Hydrometers by Robert Frease and Brian Christman, The Volunteer Monitor, Spring 1993).

I graphed the data with a scatter plot and calculated linear regressions using Excel software. I decided to calculate the mainstem and creek regressions separately because the creek stations were much closer to freshwater inflow, so salinities near zero were more common in the creeks. The scatter plot for the mainstem sites is shown at right. (For the sake of simplicity, only the results from the mainstem stations are included in this article. Readers interested in the creek results may contact me at the address below.)

If the two methods gave identical results, all the data points would fall right on the dashed line shown on the graph (the line y = x). In fact, though, most of the data points fall above this line, indicating that the hydrometer salinity values were consistently higher than the conductivity salinity values. This difference was largest at low salinity. The difference was probably due to suspended solids (and possibly non-conductive dissolved solids), which would raise the hydrometer reading (since it measures specific gravity) but not the conductivity reading.

The linear regression equation for the data shown in the graph was HS = 1.9807 + 0.9257(CS)

(where HS = hydrometer salinity;
CS = conductivity salinity)

Adjusting hydrometer salinity data

The linear regression equation can be used to adjust the data-that is, to estimate what the conductivity salinity would be for a given hydrometer salinity value. This is done by algebraically rearranging the regression equation. For the Magothy River example, the rearranged equation was

CS = (HS - 1.9807)/0.9257

Plugging in the hydrometer reading (HS) yields the estimated conductivity reading (CS). When the adjusted value was less than zero, I set it to zero. This adjustment lowered the Magothy River hydrometer data by an average of 1.3 ppt (range 0.6 to 1.8 ppt). Salinity differences of this magnitude could be important if sessile plants or animals, such as SAV or oysters, were near the limit of their salinity tolerance.

I used my adjusted hydrometer salinity data for a comparison with data from the Magothy station monitored by the Maryland Department of Natural Resources (DNR), which is being done by DNR to see if there are consistent differences in water quality due to station location. DNR wants to find out whether nearshore water quality for living resource growth can be assessed using water quality data collected at mid-channel sites. I am also using adjusted hydrometer data to help select areas for SAV restoration projects in the Magothy. If your volunteer monitoring group wants to compare salinity data measured by another method to conductivity data, I recommend that you conduct the same type of study that I did. Start by collecting (or-if you can-convincing someone else to collect) a series of paired comparison samples, one with your method and one with a conductivity meter, at the same stations. When you have collected enough data (at least 80 100 pairs over the normal range of salin-ities encountered, depending on variability), you can calculate a regression for your water body.

One word of caution: Groups using a hydrometer might be tempted to simply plug in their hydrometer readings to the Magothy River equation given above. However, I do not recommend this. Each water body will have a different equation, which must be experimentally determined. (For example, the creek data I collected yielded a different regression equation than the mainstem data.)

If any readers have salinity method comparison data, please contact me at the address below. I have also posted a request for comparison data sets (for salinity or other methods) on EPA s Surf Your Watershed Web page. To respond, go to http://www.epa.gov/surf, click on Speak Out! and View by Category, then click on Volunteer Monitoring. Peter Bergstrom is the volunteer monitoring coordinator of the Magothy River Association, and a biologist with U.S. Fish & Wildlife Service, 177 Admiral Cochrane Drive, Annapolis, MD 21401; 410/573-4554; email peter_bergstrom@mail.fws.gov.

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Conductivity Meters

Even though conductivity meters have become more affordable, they are still too expensive for most volunteer monitoring programs. The least expensive"pocket" conductivity meters cost about $60. These generally have ATC (automatic temperature compensation) and read conductivity either in milliS/cm (mS/cm) or microS/cm (µS/cm) at 25°C. (The unit S, or siemens, is a unit of electrical resistance and is identical to the mho-which by the way is ohm spelled backwards. The symbol µ is the Greek letter mu and stands for"micro."[1])

The highest range currently available in pocket meters is up to 19.9 mS/cm, or about 12 ppt salinity. The range can be extended by dilution, but this must be done carefully. Larger meters with higher ranges (up to 75-100 mS/cm) start at about $250.

You can use a table or chart to convert conductivity readings to salinity in parts per thousand (ppt). Alternatively, if you have a computer and a spreadsheet you can use the following equation, where C = conductivity in mS/cm at 25°C and K is a constant (32.188). (This equation is built into the Hydrolab meters used by many professional monitors.)

Salinity (ppt) = $20 + 0.69608 (C-K) + 0.0013094 (C-K)^2 - 0.000011918 (C-K)^3 + 0.00000017392 (C-K)4 - 0.0000000031112 (C-K)5$

In your spreadsheet, the formula should be typed in using the following format to calculate salinity in the current cell. (This example uses Excel syntax. It assumes that conductivity is in column B and the current cell is in row 4.)

=20 + 0.69608*(B4-32.188) + 0.0013094*(B4-32.188)^2- 1.1918E-05*(B4-32.188)^3 + 1.7392E-07*(B4-32.188)^4- 3.1112E-09*(B4-32.188)^5 (where * is multiplication, ^ is exponentiation, and E-05 means"times 10-5)

[1] Editor s note: Careful readers of Steve Wildberger s 1993 article will notice that"micromho and"microS are erroneously abbreviated as"mmho" and"mS", rather than"µmho and"µS. This error occurred when the desktop publishing software"helpfully" decided to convert all the µ s to m s (apparently the program doesn t speak Greek). My apologies for not catching this glitch, and for any confusion that may have been caused.

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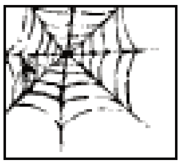

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National Conference Proceedings

The Proceedings for the fifth National Volunteer Monitoring Conference will be available in August 1997. The conference, which was held last summer at the University of Wisconsin"Madison, focused on the theme of "promoting watershed stewardship."

The 120-page Proceedings contains the full text of the plenary address by Gaylord Nelson, former governor of Wisconsin and the founder of Earth Day, plus papers from 64 conference presentations and workshops. You'll find reports on topics ranging from "Making Stewardship Measurable" to "Media Strategies for Cheapskates" to "Watershed Ecology and Indicators."

In addition, the Proceedings contains summary reports from a number of special discussion sessions. Some of these focused on a particular topic, such as "Working with Youth Groups" or "Monitoring Rivers and Streams." Others were regional breakout sessions in which attendees met to brainstorm about needs, goals, and strategies for their region.

Everyone who attended the conference will automatically receive a copy of the Proceedings. The Proceedings will also be available at no charge from Alice Mayio, Volunteer Monitoring Coordinator, U.S. EPA, 4503F, Washington, DC 20460; ph. 202/260-7018; fax 202/260-1977.

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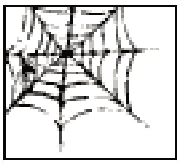

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Volunteer Monitoring on the Web

Ken Cooke, Kentucky Water Watch coordinator and Web master extraordinaire, recently ran a query on an online search service for the term "water quality" and came up with over 30,000 Web pages. Narrowing the search to "volunteer monitoring" still yielded over 2,000 sites.

Don't have time to sort through 2,000 sites? Not to worry-Ken has done the work for you. He has created a special Web page that lists over 40 high-quality sites dealing directly with volunteer monitoring. (For those not content with just 40 sites, the page includes a link to another Kentucky Water Watch Web site, "Links We Like," which contains over 100 additional water-related sites.) Each entry on the volunteer monitoring page is "hot-linked" to the corresponding Web site, so all you have to do is click on the name to get directly to the site. To find KenÕs list, point your Web browser to http://www.state.ky.us/nrepc/water/vm.htm. From there youÕre just a mouse click away from such sites as:

- EPA's volunteer monitoring sites
- International, national, state, and local monitoring organizations
- The Great American Secchi Dip-In
- The Volunteer Monitor newsletter
- National Directory of Citizen Volunteer Monitoring Programs
- K-12 monitoring programs
- Online data entry and retrieval systems
- Online macroinvertebrate key

If you have a volunteer monitoring site that is not included, send an email message to kywwp@igc.org to be added.

The following sites are of special interest to volunteer monitors:

- **EPA's Surf Your Watershed** (http://www.epa.gov/surf) allows users to find and share environmental information and maps. Search by topic or watershed, add information on your own watershed, or use the "Speak Out" section to communicate with other users.
- **EPA's Watershed Tools**(http://www.epa.gov/OWOW/watershed/tools/) contains descriptions of several hundred watershed management tools-methods for data collection, modeling, community outreach, training, etc. (Also available as a printed document from NCEPI, 11029 Kenwood Rd., Bldg. 5, Cincinnati, OH 45242; 513/489-8695.)
- U.S. Geological SurveyÕs **Water Resources of the U.S.** (http://water.usgs.gov/) includes real-time data (such as stream flows) from USGS gaging stations, plus other water quality information and a list of publications.

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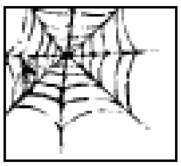

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CASE STUDY: CHEMICAL TESTING

Coyote Creek Riparian Station

During the pilot year of Coyote Creek Riparian Station's creek monitoring program in Santa Clara County, CA, parallel testing was performed to evaluate both the program protocols and the volunteers' performance. The study was conducted by Theresa Rigney as part of her Master's thesis research, and sponsored by the Regional Water Quality Control Board (RWQCB), which provided lab analyses.

Study design:

On 10 separate occasions over a 1-year period, Rigney accompanied the volunteers and conducted parallel testing, using a YSI meter for dissolved oxygen, temperature, pH, and conductivity; a turbidity meter for turbidity; and laboratory analysis (at RWQCB lab) for ammonia, nitrate, nitrite, and phosphate. Rigney's results were compared with results obtained by volunteers using field kits.

Results were analyzed by calculating mean, standard deviation, range, t-test (two-tailed), and linear regression.

Outcomes

- For temperature, DO, pH, conductivity, and turbidity, the volunteers' results were found to be within acceptable limits.

- For nutrients (ammonia, nitrate, nitrite, and phosphorus), the volunteers' results were not within acceptable limits. This was judged to be due to the limitations of the field kits, and these parameters were dropped from future testing.

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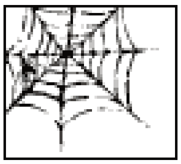

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Parallel Testing - Volunteers Versus Professionals

In the last issue of The Volunteer Monitor we invited readers to submit examples of parallel or "side by side" testing that compared volunteer monitors to professionals. Some of the responses are profiled as "case studies" on this and the following pages.

The examples came from a wide variety of volunteer monitoring programs-large and small; long-established or just starting up; programs that work primarily with adult volunteers and those that work with schools. These groups were also engaged in a wide spectrum of monitoring activities, so it's not surprising that there were some differences in their reasons for conducting side-by-side testing, in their study designs, and in their criteria for interpreting the results.

Why do parallel testing?

Often a prime motivation for conducting parallel testing is to assure government agencies that volunteers' data are reliable enough for the agencies to use. The examples we received indicate that side-by-side studies can indeed be very effective in accomplishing this goal. Sandy Fisher, Program Director for Florida Lakewatch, says that the results of that program's side-by-side testing changed the attitude of Florida officials "180 degrees"-to the point where she now gets hundreds of calls requesting the volunteers' data.

But an equally (or more) important reason to do parallel testing is for the program's own internal use. Such testing helps staff and volunteers answer the basic question, How are we doing? It shows where the program's strong and weak points are. In fact, one of the most encouraging lessons to emerge from the examples submitted to The Volunteer Monitor is that all the programs benefited greatly from their side-by-side studies-regardless of how "good" the results were in the sense of how closely the volunteers matched the professionals.

To begin with, the simple fact that the study is being conducted has an immediate positive effect. Volunteers appreciate the effort being devoted to assuring the quality of their data; it lets them know that their work is taken seriously. For students, there are educational benefits as well. Adêle Ho, NPDES coordinator for the City of San Pablo, helped oversee a program in which elementary and high school students monitored Wildcat Creek. Ho says, "When scientists joined students at the creek to conduct parallel testing, the students had a chance to observe scientists at work. The scientists also came into the classroom and talked about scientific data collection."

Most useful of all, parallel testing helps programs spot problems and make improvements. Often groups learn as much about their methods as their volunteers' technique-the volunteers may do everything right, but a problem with a method is uncovered. Indeed, in almost every example sent in the program had used parallel testing results to improve training, methodology, or manuals.

- Illinois River Watch Network discovered that volunteers were having trouble distinguishing bloodworms and non-bloodworm midges. The problem was resolved by changing the descriptions in the key.
- University of Rhode Island Water Watch found a high degree of sample-to-sample variability for chlorophyll results, even for staff-collected duplicate samples. To take into account the inherent patchiness of algae, the program changed its protocols so that volunteers now routinely collect two samples for chlorophyll.
- Connecticut River Watch volunteer protocols initially called for a 2-minute timed collection for sampling macroinvertebrates. When side-by-side testing revealed a low macroinvertebrate density in volunteer samples compared to the professionally collected data, the program switched to a more thorough, untimed collection.
- After a year of side-by-side testing for its pilot monitoring project, Coyote Creek Riparian Station concluded that the precision and accuracy of field kits for nutrients were unacceptable for the program's needs, so these tests were dropped.

How close is close enough?

How do you evaluate the results of parallel testing? How do you decide whether your volunteers' results are "close enough"? The answers go right back to your reasons for doing the monitoring in the first place. A group that is monitoring for educational purposes or to raise "red flags" generally will not need as high a level of data quality as a group that wants to detect trends or pinpoint the cause of a specific problem. Groups may also set different target levels of accuracy for different parameters; these could depend both on the uses of the data and the variability of the measurements. For example, it would be realistic to expect a greater level of accuracy with temperature measurements than with fecal coliform counts.

Once you have decided on a target accuracy level-say, you decide that for a certain parameter you want your volunteer results to be within plus or minus 10% of the professional results-you may want to calculate some simple statistics to help determine whether the data meet that target. For some pointers on using statistics, please see "Tips for Statistical Analyses of Parallel Studies" on the following page.

Be realistic in setting goals for data quality; there's no point wasting a lot of effort struggling for a higher level of quality than you really need. "I hate to see volunteer monitors chasing the chalice of perfect data," says Meg Kerr, a member of The Volunteer Monitor editorial board. "Getting high quality data has a very high cost attached to it. Groups should ask themselves whether this is the best use of time and resources." Kerr points out that many volunteer monitoring "success stories"-instances where problems were identified and solved-did not depend on the highest quality data, and adds, "Just having volunteers out looking at conditions is valuable."

When monitoring is conducted by school classes, education is often the most important goal. Project SEARCH, a program that works with nearly 100 high schools in Connecticut, does extensive parallel testing of the students' results, but the emphasis is more on identifying problems and improving the program than on obtaining data of the highest quality. If the students "flunk" the target accuracy levels for some parameters, staff members are philosophical. "Project SEARCH works with all kinds of students, from learning-disabled to honors students," says Mike Beauchene, the program's Water Resource Biologist. "It would be unrealistic to expect consistently high quality data. When the students' results match the professional lab's, we jump up and down. If they are far apart, we ask why and look for ways to solve the problem." (Note: For more on SEARCH, see "Wanted-Preferably Alive," on page 1 of this issue.)

Many thanks to Denise Stoeckel and Woodrow Setzer for reviewing all the parallel testing examples and providing valuable insights.

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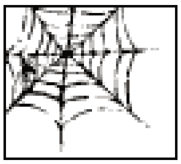

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CASE STUDIES: LAKE MONITORING

University of Rhode Island Watershed Watch

Over 200 volunteers, mainly adults, monitor lakes in Rhode Island. In 1992, the program obtained a federal Clean Lakes grant to conduct parallel testing of volunteers versus program staff. The primary purpose of the study was to convince state agencies that URIWW data were credible, so they would use the data.

Study design: URIWW staff member Elizabeth Herron accompanied volunteers at 21 lakes on their regular sampling date. The volunteer performed sampling as usual. Herron collected one set of samples using the same methods and equipment as the volunteer, plus additional samples using EPA Clean Lakes methods. This protocol allowed evaluation of both (a) volunteer performance and (b) URIWW methodology as compared to EPA Clean Lakes methodology.

Methods assessed were: Secchi reading; dissolved oxygen; sample filtration for chlorophyll; and sample collection (for later analysis at URI lab).

The data were analyzed using mean difference, standard deviation of the mean difference, standard error of the mean difference, 95% confidence interval of the mean difference, percent error, quartiles, range, and regression.

Outcomes

- No statistically significant differences were found between samples collected by volunteers, samples collected by staff according to URIWW protocol, and samples collected by staff according to EPA protocol for any parameter.
- The study proved to the state that volunteers were doing good work. The state now uses the volunteers' data to help identify priorities and establish standards, and incorporates volunteer results into the 305(b) report (the biennial water quality report that states submit to EPA).

Florida Lakewatch

Adult volunteers on about 600 Florida lakes take Secchi readings, filter samples for chlorophyll analysis, and collect grab samples for nitrogen and phosphorus (analyses are performed at the Lakewatch lab.)

Study design: Twice during 1991, parallel testing was conducted on 125 lakes at the request of the Florida Department of Environmental Protection. Biologists accompanied volunteers on their regular sampling trip and tested the same parameters as the volunteers, as well as collecting additional samples for other purposes. The study was "blind"-that is, volunteers did not know that they were being tested.

Outcomes

- There was no statistically significant difference in the values obtained for any of the parameters tested.
- As a result of the study, Fisher says, "the attitude of Florida officials changed 180 degrees. Now I get 600 requests per year for our data-from regulatory agencies, city planners, lake managers, professional consulting firms, and citizen groups."

Metropolitan Council Citizen-Assisted Monitoring Program

Adult volunteers in east central Minnesota monitor lakes biweekly, measuring temperature and transparency (Secchi) and collecting surface water samples which are analyzed at the Metropolitan Council lab for phosphorus, nitrogen, and chlorophyll. One of the program's goals is for the volunteer data to be as reliable and credible as data collected by the Council's professional staff.

Study design: Parallel testing is a regular, ongoing component of the program's quality assurance. A council staff member monitors the same site, using same type of equipment and same methods as volunteers. During the 1996 monitoring season, 41 of the 53 sites were checked by staff. Results are analyzed using the paired t-test and regression analysis.

Outcome

- There was excellent agreement between Council staff and volunteer results for all parameters.

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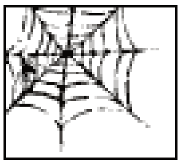

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CASE STUDIES: MACROINVERTEBRATE MONITORING

Illinois RiverWatch Network

Volunteers (all adults) do habitat surveys, then collect and preserve macroinvertebrate specimens which they later identify in a lab, usually to family level.

Study design: Parallel testing is conducted as an ongoing part of the quality assurance program. Using a random process, Denise Stoeckel (the project's quality assurance officer) selects 30% of the volunteer sites each year. For each selected site, she re-examines the preserved specimen to verify the volunteer's identification. The target level of accuracy for each taxon is correct identification in at least 90% of the samples.

Stoeckel points out that this design does not constitute complete parallel testing, since she doesn't replicate the volunteers' collection procedures. This year, Stoeckel plans to "shadow" a few volunteers to do a side-by-side evaluation of the whole monitoring process, including habitat assessment and sample collection.

Outcomes

- In 1995, the target level of accuracy was met for 30 out of the 33 taxa identified (6 taxa were correctly identified in all samples and 24 were correctly identified in 90% or more of samples).
- When biotic indexes were recalculated based on corrected identifications, the stream ratings did not change.

- For taxa that did not meet target accuracy levels, the key was revised to make identification easier.

Connecticut RiverWatch

Volunteers, both adults and high school students, monitor 40 sites on the lower Connecticut River and its tributaries. As with IL RWN (above) macroinvertebrates are preserved and later identified to family level.

Study design: Once a year, starting in 1992, CT Department of Environmental Protection aquatic invertebrate specialist Guy Hoffman has collected benthic macroinvertebrate samples at one RiverWatch site within a few days of the volunteer team's collection. (Note that this study design tests the volunteers' performance for both collection and identification.)

Outcomes

- Based on the first year's parallel testing results, the program modified the volunteers' collection protocol.
- Each year the volunteer team and the DEP biologist have found the same taxa (though not always the same percentages). Both agree that the most pollution-sensitive organisms are absent, and both obtain similar overall stream ratings based on biotic index.

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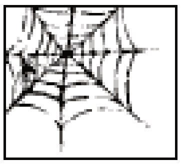

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Tips for Statistical Analyses of Parallel Studies

by Woodrow Setzer

Editor's note: Woodrow Setzer (who is both a volunteer river monitor and a professional biostatistician) reviewed all the examples of parallel testing that were submitted to The Volunteer Monitor, *then contributed the following advice on using statistics to analyze such studies. Setzer hopes to cover the topic of statistical testing more thoroughly in a future issue of* The Volunteer Monitor.)

The following brief pointers about statistical analysis are, obviously, not a thorough treatment of the topic. Those who wish to learn more are encouraged to find a good book about statistics for biological applications; one such is Biometry, third edition, by R. R. Sokal and F. J. Rohlf, Freeman Press. (Computer software manuals, unfortunately, are rarely a good place to learn about statistics.) In the end, though, there is really no substitute for familiarity with statistical methodology-so don t be shy about searching for people with professional training in statistics to help out with problems.

Plan ahead

Plan in the statistical analysis from the start. An important part of planning is to state very clearly and precisely exactly what questions you are asking, and how the statistical analysis will answer those questions. With parallel testing (professional versus volunteer results), the basic question is, How closely do the volunteer results match the professional results? You should also consider how precisely you want to be able to answer that question; this in turn will help to determine the number of samples you need to take.

Use common sense

Look at simple summaries and plots of the data before you carry out more complicated statistical manipulations. Two problems to look for are individual values that are very different from all the others (possibly due to a typographic error), and differences whose absolute magnitudes increase as the average measurement increases. Both of these situations may make the variability seem bigger than it really is.

In the first case, you would certainly want to go back over all the data records to see if there had been a transcription error, and you might want to calculate statistics two ways, first with and then without the offending observation. In the second case, you might want to work with a logarithmic transformation of the original data.

A good way to show up both of these problems is to plot the differences between each pair versus the means of each pair. For example, suppose you are comparing 20 pairs of dissolved oxygen data. Each pair consists of one volunteer measurement (V) and one professional measurement (P). Plot the differences, V P, on the y-axis, versus the means, (V P)/2, on the x-axis. If one y-value stands out above (or below) the rest by a great deal, you have a problem of the first kind, and if the range of the y-values changes from left to right, you have a problem of the second kind.

Your *t*-test isn t significant-How happy should you be?

In the typical paired design used in most volunteer monitoring parallel studies, the most important statistic to estimate is the difference between the volunteers results and a gold standard (typically, results obtained by a water quality professional). The second most important statistic is a measure of the precision of that first statistic, such as a standard error or a confidence interval (confidence intervals are preferable).

In many of the examples sent in to The Volunteer Monitor, volunteer monitoring groups calculated that important first statistic-the difference-and then went on to do a t-test or other statistical test to tell them whether that difference was significant. Often there is really no need to conduct such a test to evaluate the results of a side-by-side study. Simply calculating the mean difference and the confidence interval on the mean difference, then comparing the results to your target level of accuracy, will usually tell you all you need to know. In fact, a statistical test such as a t-test can be downright misleading, especially if you don t also look at confidence intervals. To understand why, we need to look at what statistical tests really do (and don t do).

Statistical tests, like the paired t-test or the Wilcoxon signed ranks test, test the null hypothesis that there is NO difference between the samples-i.e., the difference is zero. The alternative hypothesis is that there is SOME difference between the samples. When the P-value for the test is smaller than some pre-defined level, for example 0.05, we say that the test rejects the null hypothesis (in other words, the difference is not zero) and the difference is statistically significant. In the case of parallel testing, you don t want to see a statistically significant difference. So if you get a big value for P, meaning you cannot reject the null hypothesis, you're happy.

However, what statistical tests alone do not tell you is how happy you should be. For that you need to calculate the confidence interval (or, alternatively, the standard error). For example, suppose you are comparing volunteers chlorophyll results to a gold standard. Let s call the volunteer results V and the professional results G, for gold. You find that the mean of G is 25 and the mean difference (V G) is 3 (for a 12% error; i.e., 3 divided by 25). You perform a statistical test, say a paired t-test, and find that the difference is not significant. At this point you would probably be tempted to believe that your volunteers are doing very well. Hold on, though. Now you calculate the 95% confidence interval for the difference and find that it is 4 to 10. This is a relatively large confidence interval. Large confidence intervals can occur if there is a lot of variability in the data (such as you might get with chlorophyll, due to the patchiness of algae), or if the sample size is small, or for other reasons. This confidence interval tells you that the percent error is somewhere in the range of 16% to 40%. If your target level of accuracy was less than a 15% error, you would certainly not be happy with this result.

In fact, you would be happier with a difference that is statistically significant but has a small confidence interval. Let s consider another example. This time you re testing dissolved oxygen, so there is less variability in the data, and you have a larger number of data points. Once again you calculate a mean for G of 25 and a mean difference (V G) of 3. You do a t-test and find that this difference is statistically significant. This time, though, you calculate a confidence interval for the difference of 2.5 to 3.5 (corresponding to a percent error of 10% to 14%). You can be reasonably confident that the true error is between 10% and 14%. Since you have decided you can tolerate an error up to 15%, you can be happy with this result.

What R^2 won't tell you

Before using a statistical procedure, make sure you know what questions it is designed to answer. For example, several of the case studies used linear regression to compare their volunteer samples to a gold standard. If the regression analysis yielded a large R^2 they were happy. But R^2 does not answer the question, How well do the x and y values match? It only answers the question, Is there a linear relationship between x and y? A large R^2 only tells you that there is a strong linear relationship-it doesn't tell you where that line is. In the hypothetical example illustrated in the graph below, the x values (professional results) and y values (volunteer results) line up very nicely, as indicated by the R^2 of 0.99. But you probably wouldn t be happy with these results.

The regression equation, which will be in the form $y = a + bx$, will give you much more useful information than the R^2 value. If the volunteer results perfectly matched the professional results, the intercept, a, would be 0, and the slope, b, would be 1 (that is, the data points would fall along the line $y = x$). To see how well the volunteer results match the professional results, look for a slope close to 1 and an intercept close to zero.

Woodrow Setzer *is a volunteer monitor with the Haw River Assembly Riverwatch project and a biostatistician in the National Health and Environmental Effects Research Lab of the U.S. EPA in Research Triangle Park, NC. He may be contacted at* wsetzer@mindspring.com.

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Watershed Activity Packet

An activities packet produced by Water Action Volunteers (WAV) provides instructions for eight simple, hands-on projects suitable for classrooms or community groups. The packet tells how to organize activities such as a stream walk, stream cleanup, "critter search," and storm drain stenciling, and also provides instructions for building simple, low-cost educational models such as "Erosion in a Bottle" and "Watershed in a Box" (see below).

WAV is a cooperative program between the University of Wisconsin Cooperative Extension and the Wisconsin Department of Natural Resources. The WAV packet is free to Wisconsin residents and $10 for nonresidents.
To order, please contact: Pamela Packer,
WAV Coordinator, WI DNR,
Bureau of Watershed Management,
101 S. Webster St., Box 7921,
Madison, WI 53707;
608/264-8948;
email ppacker @facstaff.wisc.edu.

Watershed in a Box

Note: This abbreviated version of the "Watershed in a Box" activity was adapted with permission from the WAV packet. Please refer to the WAV packet for more detailed instructions, as well as background material on nonpoint source pollution.

This extremely simple watershed model, which can be constructed in a few minutes, demonstrates how different types of nonpoint source pollution can affect a stream.

1. **Get a box.** Use a box cover or shallow box, 12" by 12" or larger.
2. **Create land forms.** Arrange pieces of foam, Styrofoam, or crumpled paper to represent hills and land forms. The highest points should be near the box wall. Leave a gully in the middle to represent a stream.
3. **Cover the land forms.** Use heavy-duty aluminum foil, shiny side up. Start in the middle and work outward, gently pressing the foil into all the hills and valleys. Fold the foil over the edge of the box. Be careful not to tear the foil.
4. **Create a community.** Use a permanent marker to draw the outline of the stream. Then draw houses, roads, farm fields, feed lots, stores, parking lots, shopping malls, factories, etc.
5. **Create a water body.** Pour a little water into the stream.
6. **Add some pollution.** Sprinkle different colors of powdered drink mix onto the model to represent different kinds of pollution. For example:
   - Red = yard care chemicals. Sprinkle around houses.
   - Green = automobile waste (e.g., oil, brake fluid, brake pad and tire wear) or road salt. Sprinkle on roadways or parking lots.
   - Brown = exposed soil. Sprinkle on construction sites or farms.
   - Blue = animal waste. Leave little piles of powder near homes and farms.
7. **Make it rain.** Use a spray bottle to make a "rain storm." Watch what happens.
8. **Discuss what you observed.** What could be done to reduce runoff pollution in the watershed?

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GREEN Public Workshops

Public workshops on three environmental education topics are being offered at various locations around the country by GREEN (Global Rivers Environmental Education Network). Each workshop lasts three days; the cost of $90 includes curriculum materials. Workshop topics are:

- **What's GREEN & WET? An Introduction to Watershed Education.** GREEN and Project WET combined forces to develop this workshop, which covers water quality testing, data analysis, and techniques for solving environmental problems.
- **Environmental Education for Empowerment: Students Solving Problems in Their Own Neighborhood.** Participants learn techniques for helping students investigate real-world problems, make decisions, and take action.
- **River of Words: Exploring Watersheds Through Poetry, Art, and Ecology.** This workshop, offered by GREEN and International Rivers Network, explores the use of poetry, art, and environmental science to investigate watersheds and take action to improve them.

For additional information, including a complete listing of workshop locations and dates, please contact: Carolyn Henne, at
GREEN, 206 S. 5th Ave.,
Suite 150, Ann Arbor,
MI 48104;
313/761-8142; (fax) 313/761-4951;
chenne@green.org.

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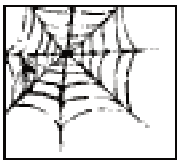

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Speak Out! About Volunteer Monitoring

EPA's Surf Your Watershed, a Web site designed to help citizens locate, use, and share environmental information on their watersheds and communities, has a new chat room service called Speak Out! When you visit Speak Out! you'll be able to participate in public discussions on volunteer monitoring topics or make private comments to others in the chat room. Other categories of subjects in Speak Out! include nonpoint sources, watershed lessons learned, and biological assessment.

To visit Speak Out! go first to the Surf Your Watershed site at http://www.epa.gov/surf and click on Speak Out! You'll be asked to register as a chat room user; in about 15 minutes, you'll be ready to go. Speak Out! is in its early stages right now-your input is needed to make this a useful tool for information sharing.

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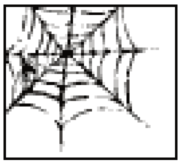

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RESOURCES

IWLA Wetland Manual

A new 235-page handbook from the Izaak Walton League of America is designed to help people become wetland stewards. Save Our Streams Handbook for Wetlands Conservation and Sustainability describes unique wetland features (soil types, hydrology, plant species), explains wetland functions and values, and includes various techniques for monitoring and protecting wetlands. The book is available for $18 from IWLA, Save Our Streams Program, 707 Conservation Lane, Gaithersburg, MD 20878-2983; 800/BUG-IWLA.

Science Project Guide

Need ideas for a science fair project? The Izaak Walton League of America's Science Project Guide for Students shows students how to use stream monitoring as a basis for a science project. The guide covers basic stream survey techniques, ideas for presenting the project, and actions students can take to help the stream. It is available for $3 from IWLA (see address above).

Starting a Watershed Organization

River NetworkÕs Starting Up: A Handbook for New River and Watershed Organizations offers advice on topics ranging from choosing a name and recruiting a board of directors to fundraising and working with the media. Available for $25 ($10 for River Network Partners) from River Network, P.O. Box 8787, Portland, OR 97207; 503/241-3506; rivernet@igc.apc.org.

Hach Co. Newsletter

Hach Company's free quarterly newsletter, News and Notes for the Analyst, contains a regular feature called "Quality Corner" that discusses quality assurance issues, including how to calculate and interpret basic statistics. Available from Hach Co., P.O. Box 369, Loveland, CO 80539; 800/227-4224.

Sourcebook from GREEN

GREEN's new Sourcebook for Watershed Education provides the framework and tools for organizing a self-sustaining watershed education program. It also offers curricula, lessons, and activities. Available for $29.95 from GREEN, 206 Fifth Ave., Suite 150, Ann Arbor, MI 48104; 313/761-8142.

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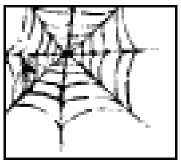

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Survey for National Volunteer Monitoring Directory

To assure your program's inclusion in the printed Directory, please return to:

Volunteer Monitoring Survey
2765 16th St,
San Francisco, CA 94103
Questions? Call (415)255-8049

Name of person completing questionnaire:___________________

Monitoring program name, exactly as you want it listed in the Directory. **Note:** Programs will be listed alphabetically. Use the first line below for the name most people will look for-e.g., if "Friends of Fox Lake" has a monitoring project called "Citizen Watch," they should list Friends of Fox Lake on the first line and Citizen Watch on the second line.

Affiliation, if you are part of a national, statewide, or regional network (e.g., Izaak Walton League, Texas Watch):

Mailing address:

phone:

fax:

e-mail:

Web site:

Monitoring program coordinator(s):

Does your program serve as an "umbrella" organization for smaller monitoring groups? Y N

NOTE: The questions below refer only to the portion of your program devoted to volunteer monitoring.

Active volunteers (excluding school classes): ________

For programs that work with schools: # Teachers: _______ # Students: _______

Approx. annual monitoring budget: $____________ Year monitoring began: 19_____

Sources of funding or in-kind support:

___ fed. gov't ___ state gov't ___ local gov't ___ foundations ___ businesses ___ memberships

___ donations ___ grassroots fundraising (events, solicitations, etc.) ___ other:______

Does program have a written QA (quality assurance) plan? Y N Is it state-approved? Y N EPA-approved? Y N

Does program have monitoring-related publications you are willing to share with, or sell to, other groups? Y N

Counties in which you monitor. This information will be used to locate your monitoring activities in EPA's "Surf Your Watershed" Web site. Please list ALL counties in which you monitor, by both county name and state (attach extra sheet if needed).

county	state	county	state	county	state

Program description. Please tell us what you would most like people to know about your program (e.g., water bodies and watersheds monitored; major monitoring projects and related activities; international projects). Space is limited! Please be brief!

Environments monitored

___ groundwater ___ river/stream ___ estuary ___ coral reef ___ marine ___ air ___ land

___ lake/pond ___ reservoir ___ beach ___ wetland ___ other:

Physical/chemical monitoring

___ water temp. ___ nitrogen ___ dissolved oxygen ___ metals ___ flow/water level

___ rainfall ___ phosphorus ___ BOD ___ hydrocarbons

___ pH ___ TSS/TDS ___ Secchi transparency ___ pesticides

___ hardness ___ conductivity ___ turbidity ___ toxicity

___ salinity ___ chloride ___ alkalinity ___ other: ________________________________

Biological monitoring

___ macroinvertebrates ___ chlorophyll ___ bacteria ___ wildlife

___ fish ___ aquatic vegetation ___ shellfish ___ exotic/invasive species (specify which: ________________)

___ habitat assessments ___ terrestrial vegetation ___ birds ___ other:

Other activities

___ debris cleanup ___ pipe surveys ___ stream channel morphology ___ construction site inspections

___ debris monitoring ___ photographic surveys ___ restoration (e.g.: __________________________)

___ land use surveys ___ human use surveys ___ storm drain stenciling ___ other:

Data uses and users:

Please indicate who uses your data, and for what.

Data users:

Community organizations University Our program (non-gov't) Local gov't State gov't Federal gov't scientists

Data Uses:

Education

Advocacy

Research

Community organizing

Screen for problems

Establish baseline conditions

Nonpoint source assessment

BMP evaluation

Land use decisions

Watershed planning

Plan restoration projects

Enforcement

Legislation

Shellfish bed closures

Swimming advisories

State 305(b) report

Other (please specify)